

Biological Sampling

Focus Questions:

- What are the biological sampling priorities for fish?
- Which fish should viabilities be collected from?
- What biological information is collected from fish?
- What physical specimens are collected from fish?

Chapter Outline

- I. Introduction
- II. Data Collection Priorities
- III. Biological Specimens Sampling
- IV. Data Collection Procedures
- V. Data Collection Forms

I. Introduction

Observer programs provide an excellent way to collect biological specimen information for use by fisheries biologists and stock assessment analysts. Observers in the WCGOP are asked to collect fish lengths, weights, sexes, otoliths, viabilities, tags, and scales in an effort to improve understanding of various fish stocks.

Fish lengths, sexes and otoliths are used to determine the relative abundance of fish year classes and the occurrence of the various year classes in commercial fishing landings. These specimen collections are also used to estimate the sexual composition of fish year classes, determine differential growth rates between sexes, and to provide length to age ratios for use in stock assessments.

Pacific halibut viabilities (injury data) are used to assess the mortality rate of Pacific halibut due to commercial fishing. The injury data collected by Observers are analyzed by staff from the International Pacific Halibut Commission (IPHC) and used to estimate yearly mortality rates.

Information from tagged fish and crabs are used by a variety of educational institutions, state agencies, and federal agencies. Fish are tagged to study fish migration, stock separation, fishing related mortality, and population dynamics. Data from tagged fish is vital to the success of these studies.

Scales are collected from salmon to verify species identification. Salmon are often extremely difficult to identify when caught either because of damage incurred during gear retrieval or strange coloration due to the proximity of spawning.

II. Data Collection Priorities

Due to potential data collection time constraints, Observers are asked to prioritize the collection of biological specimen information by species and information taken. In addition, biological sampling should only be undertaken after collecting comprehensive catch and species composition information. Biological Specimens are taken from DISCARD only.

Biospecimen data collection in order of priority:

1. Tagged Fish
 - Collect length, weight, sex, and tag.
 - Collect otoliths if appropriate (see details below).
2. Salmon
 - Collect length and sex.
 - Collect snout if adipose fin is missing (tagged salmon).
 - If possible, collect weight.
 - Collect scales if appropriate (see details below).
3. High Priority Rockfish Species and Lingcod
 - Collect length and sex.
 - If possible, collect weight and otoliths.
4. Pacific Halibut
 - Collect length and viability.
 - If possible, collect weight.

Data Collection from Tagged Fish

Sablefish and salmon are the most commonly encountered tagged fish. Occasionally tagged Pacific cod, Pacific halibut, California halibut, pollock, shortspine thornyhead and some rockfish species may be seen as well.

Collect the following data from tagged fish:

1. Length
2. Sex
3. Otoliths - Except for sablefish with blue spaghetti tags and salmon. In these cases, collect the entire snout.
4. Weight
5. Tag – Collect either the external tag or salmon snout.

Data Collection from Salmon

Salmon are difficult to identify correctly and may be tagged internally or externally. Watch for missing adipose fins which mark fish with internal tags. Scales are collected both to verify species identification and from all tagged salmon.

Collect the following data from salmon:

1. Length
2. Sex
3. Weight

4. Scales

- From the first five fish of each salmon species encountered during your first contract and the first two fish from each subsequent contract.
- Whenever species identification is in doubt.
- When an external tag or tagged snout is collected.

5. Snout if the adipose fin is missing.

Data Collection From High Priority Rockfish Species and Lingcod

There are eleven high priority species (mostly rockfish) on the west coast about which limited biological information has been collected to date. Stock assessment analysts have asked that WCGOP Observers focus on collecting length, sex and otolith information from these species.

Following is the list of high priority species in order of data collection priority:

1. Canary Rockfish
2. Yelloweye Rockfish
3. Bocaccio Rockfish**
4. Cowcod**
5. Pacific Ocean Perch
6. Lingcod
7. Dark-blotched Rockfish
8. Widow Rockfish
9. Rougheye Rockfish
10. Shortraker Rockfish
11. Silvergrey Rockfish

Priority only for fish collected South of 40°10' (Cape Mendocino)

Collect the following data from high priority species:

1. Length
2. Sex (except for fish being released alive).
3. Otoliths (except for lingcod, Bocaccio rockfish and rockfish species being released alive).
4. Weight if otoliths are taken.

Collect high priority fish for sampling based on the following guidelines:

1. Collect data only from discarded fish.
2. Collect fish in order of priority.
3. Collect data from all discarded high priority species in the species composition sample if possible.
4. If there is a mixture of many high priority fish in the species composition sample, collect 5 fish from each species in order of priority for a minimum of 20 fish total per haul.

Data Collection from Pacific Halibut

Pacific halibut are frequently immediately sorted from the catch and returned alive to the sea. Collect lengths and viabilities from the discarded Pacific halibut as the crew is sorting.

Collect the following data from discarded Pacific halibut:

1. Length (do not estimate).
2. Viability
3. Weight if possible.

III. Biological Specimen Sampling

Biological information is collected on individual fish for a variety of reasons. In some instances it is pertinent that the information be collected in a random fashion while in others, such as with tagged fish, information is, of necessity, collected in a non-random fashion.

There are four sample methods for biological sampling. The primary factors used to differentiate these methods are:

1. Whether the individuals used for biological sampling were within the species composition sample.
2. Whether the individuals used for biological sampling were randomly selected.

Biological Sampling Methods

Sample Method 6 – Outside and Nonrandom

- Individuals are not part of a species composition sample and have NOT been randomly selected.
- Use this method for tagged fish that have been collected opportunistically during a haul/set.

Sample Method 7 – Outside and Random

- Individuals are not part of a species composition sample and have been randomly selected.
- Use this method for Pacific halibut when lengths/viabilities have been taken for randomly selected individuals from the haul/set but there was not a species composition sample because actual weights of halibut were not obtained.

Sample Method 8 – Inside and Nonrandom

- Individuals are part of a species composition sample and have NOT been randomly selected.
- Use this method for tagged fish that have been collected opportunistically from a species composition sample.

Sample Method 9 – Inside and Random

- Individuals are part of a species composition sample and have been randomly selected.
- Use this method when taking biological data from all individuals or from randomly selected individuals of a particular species within a species composition sample.

Random Sampling

In general, individuals used for biospecimen sampling should be selected from within a species composition sample. Only on rare occasions is it necessary to create an independent random sample for biological specimens.

Random Sampling Within a Species Composition Sample

Selecting individuals for biological sampling from within a species composition sample is encouraged. When collecting individuals from inside a species composition sample, all of the individuals of a single species make up a single population (see Chapter 3 for a review of Random Sampling Theory). There are two ways that a random sample can be taken from a population.

- **All** individuals in the population are selected.
- A random subsample of the individuals in the population is selected.

Subsamples may be taken using any one of the following random sampling methods.

- Spatial – Randomly select a unit of gear or an area (portion of deck or bin, specific basket) to collect individuals from.
- Temporal – Randomly select a point in time to collect individuals.
- Systematic – Select a random start point (spatial or temporal) and take individuals at set intervals. In order to use a systematic system you must know approximately how many of the target species are in the population.

Example:

1. The crew on a trawler is sorting out a scupper and the Observer is whole hauling the discard.
2. The Observer estimates that 100 Pacific Ocean Perch (POP) will be discarded. There are no other priority species present.
3. The Observer refers to the Biological Sampling chapter in his manual and verifies that he needs to take sexed lengths from 20 of the discarded POP.
4. The Observer decides to do a systematic subsample of the POP within his species composition sample.
5. The Observer divides the estimated number of POP in the haul by the number he needs to sample ($100 / 20 = 5$). This tells him he need to collect 1 fish out of every 5.

6. The Observer randomly selects a number between 1 and 5. This will be the first POP collected. He selects 5.
7. The Observer collects the fifth POP the crew sorts and every 5th POP thereafter (5, 10, 15, 20, 25...) for biological specimen sampling.
8. The POP sexed lengths will be recorded on the Length Frequency form with a sample method of 9 – Inside and Random.

Random Sampling Outside a Species Composition Sample

Selecting individuals for biospecimen sampling from outside a species composition sample should be a rare event. Normally this type of sampling is only used in two circumstances.

- When biological information is impossible to collect during the tally sampling period.
- When weighing Pacific halibut is not possible.

If biological information is being collected from individuals that need to be released alive, it may not be possible to accurately tally sample and collect the needed individuals for biological information at the same time. In this circumstance, the only option is to collect individuals for biological specimens during a non-tally period. A random sampling frame should be designed where all selected individuals are sampled.

Often lengths and viabilities will be collected from Pacific halibut but actual weights will not be taken, instead the length /weight conversion table will be used to obtain weights. In this circumstance, the PHLB (Pacific Halibut) catch category will have a biospecimen sample without an associated species composition.

When randomly collecting individuals from outside a species composition sample use the following guidelines:

- Size the sample appropriately for the number of individuals needed.
- Select all individuals in the population.
- Do not subsample the population.

Example:

1. The Observer on a longline vessel is tallying skates 1-10 and 21-30 of a 40 skate set.
2. There are fish on almost every hook so it is impossible for the Observer to look away even for a minute to collect biological information.
3. The Observer needs to collect lengths and viabilities on Pacific halibut. Viabilities need to be taken immediately as normal crew handling is to release the fish right away and viabilities must be taken at normal point of release.
4. The Observer determines during the initial tally period that approximately 10 Pacific halibut are being caught on every skate.
5. The Observer calculates that during the non-tally periods (20 skates) 200 Pacific halibut will be caught. He wants to take lengths and viabilities from 20.
6. The Observer divides the estimated number of Pacific halibut that will be caught during the non-tally period by the number he wants to take data from ($200/20 = 10$). This tells him he needs to sample 1 fish out of every 10.
7. The Observer randomly selects a number between 1 and 10. He selects 2. This means that the second Pacific halibut caught will be his first sample fish.

8. The Observer asks the gaff-man to land the second Pacific halibut that comes up during the non tally period. He records the length and viability of this fish.
9. The Observer then asks the gaff-man to land every 10th Pacific halibut caught thereafter and takes lengths and viabilities on all of them (the population in this case is every 10th fish, beginning with fish #2).
10. The viabilities will be recorded on the Biological Specimen form with a sample method of 7 – Outside and Random.

Non-Random Sampling

The **only** time that fish should be selected in a non-random fashion is when biological information is being collected from tagged fish. Tagged fish may be taken from either inside or outside of a species composition sample.

A tagged fish has been collected non-randomly if either of the following apply:

- The tagged fish is NOT part of a species composition sample or a random sample taken from outside the species composition.
- The tagged fish is within a species composition sample but is NOT part of a random sample for biological specimens.

IV. Data Collection Procedures

Fish lengths, sexes, and otoliths are collected from a wide variety of species. While the information gathered is the same for all species, the collection procedures often vary. It is important to utilize the appropriate procedure for each species to insure accurate data collection.



Figure 6- 1: Rockfish Length

Lengthing Fish

Data Collection Guidelines

1. Collect lengths for tagged fish.
2. Collect lengths for Pacific halibut when viabilities are being taken.
3. Collect lengths for salmon.
4. Collect lengths for high priority species such as rockfish or lingcod.

Preparing to Measure Fish

Before you begin collecting fish for length measurements, set up an area to measure fish. You will need to use or create a “table” large enough to lay a fish on the plastic length strip. If there is no table present, use the NMFS aluminum board, baskets, deck bin boards or the deck as a table.

Selected species should be collected in a random fashion for length measurements and should only be collected from discarded species.

Measuring Fish

Fork length is the fish length measurement method used by the WCGOP and other NOAA Fisheries researchers. Fork

length is the length from the tip of the snout or jaw (whichever sticks out most) to the end of the middle rays of the caudal fin (See Figure 6-1 and 6-2). The only exception to this rule is grenadier length, which is measured from the snout to the insertion of the anal fin.

You will be given plastic measuring strips marked at centimeter increments. The first line printed on the strip is 4.5 cm, and the space between that line and the next line is .5 cm. Check your plastic strip on both sides to insure that the first line is actually located 4.5 cm from the end of the strip. Sometimes the manufacturer has cut the strip incorrectly. Notice that the 10-centimeter increments are not marked with a number. This is to facilitate offsetting the strip by 10, 20, or 30 centimeters for larger fish.

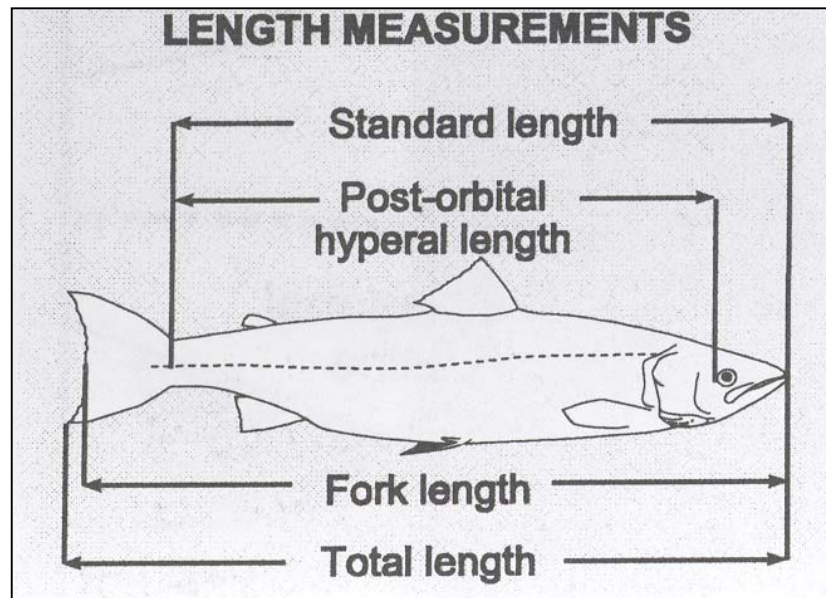


Figure 6- 2: Length Measurements

Measure fish using the following procedure:

1. Lay the fish flat on the plastic measuring strip parallel to the center-line.
2. Close the jaws.
3. Nudge the fish snout against the end of the aluminum board.
4. Stretch out the tail to find the middle rays.
5. Make a mark on the plastic measuring strip in the space where the fork length falls (above the center line for males and below the center line for females). If the fork length falls on a printed line on the strip, try re-measuring the fish first, then if the length still falls directly on the line, use the lower centimeter measurement.
6. When collecting only sex/length data transfer the number of pencil marks (frequency) made at each centimeter measurement (size group) from the plastic length strip to the Length Frequency Form.
7. When collecting individual weight information or taking scales, otoliths, snouts, or viabilities in addition to sex/length information be sure to keep all data from each individual fish together and record it on the Biospecimen Form.
8. Clean the length strip with scouring powder to remove the marks and ready it for the next haul's lengths. Do not scrub too hard because the centimeter lines will be scoured off.



Figure 6- 3: Sexing Fish

Sexing Fish

Data Collection Guidelines

1. Collect sexes from tagged fish.
2. Collect sexes when otoliths are taken.
3. Collect sexes from salmon when scales or snouts are taken.
4. Collect sexes when taking lengths from dead high priority rockfish.

DO NOT sex fish in the following situations:

1. When the fish are being discarded alive. This is common in the Live Fish and Dory Fleet fisheries.
2. When the fish is a hardy species that is likely to survive being discarded. Lingcod, sablefish and Pacific halibut are considered to be hardy species and should not be sexed unless they are dead and tagged.
3. When the fish is tagged but the vessel is retaining the fish and sexing it would damage the product (some vessel may still allow you to sex it, but usually not).

Preparing to Sex Fish

Roundfish, sablefish, rockfish, flatfish and salmon are all sexed slightly differently due to variations in anatomy (See Figures 6-3 and 6-4). Attention should be paid to the cut necessary to locate the gonads, the location of the gonads within the body cavity and the physical description of the gonads.

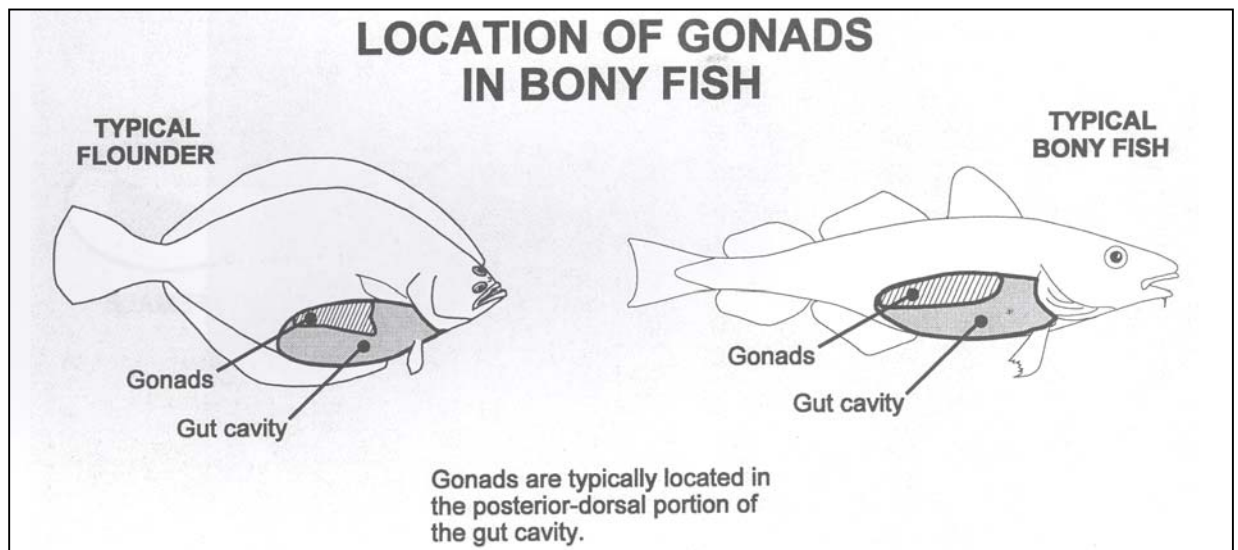


Figure 6- 4: Location of Gonads

Sexing Roundfish

Roundfish gonads are in the visceral cavity, ahead of the anus (See Figure 6-5). Insert a knife or scalpel blade in or near the anus and cut forward toward the head. There are only two organs attached directly to the anus, the intestine and the gonads. Carefully move the other organs aside to get a clear view of the tubes attached to the anus, then pull on the tubes to discern which is the intestine (coiled and ending at the stomach) and which is the gonad (ending in paired structures near the backbone).

The ovaries are paired sacs that are typically pink or orange (or clear when immature). When the ovaries are mature, you should be able to see eggs inside (the sacs will look granular). Pacific cod ovaries often have a black covering on each sac.

The testes of Gadids (cod family) look very different from ovaries. When mature, the testes are convoluted, opaque and smooth in texture. In a mature male, the testes are best described as “greasy-looking, white, twisted Ramen

noodles.” Immature testes will be pink or cream colored, have a ruffled look to the edges of the tubes, and be located near the backbone. Often, the paired gonads are fused together and look like a single structure.

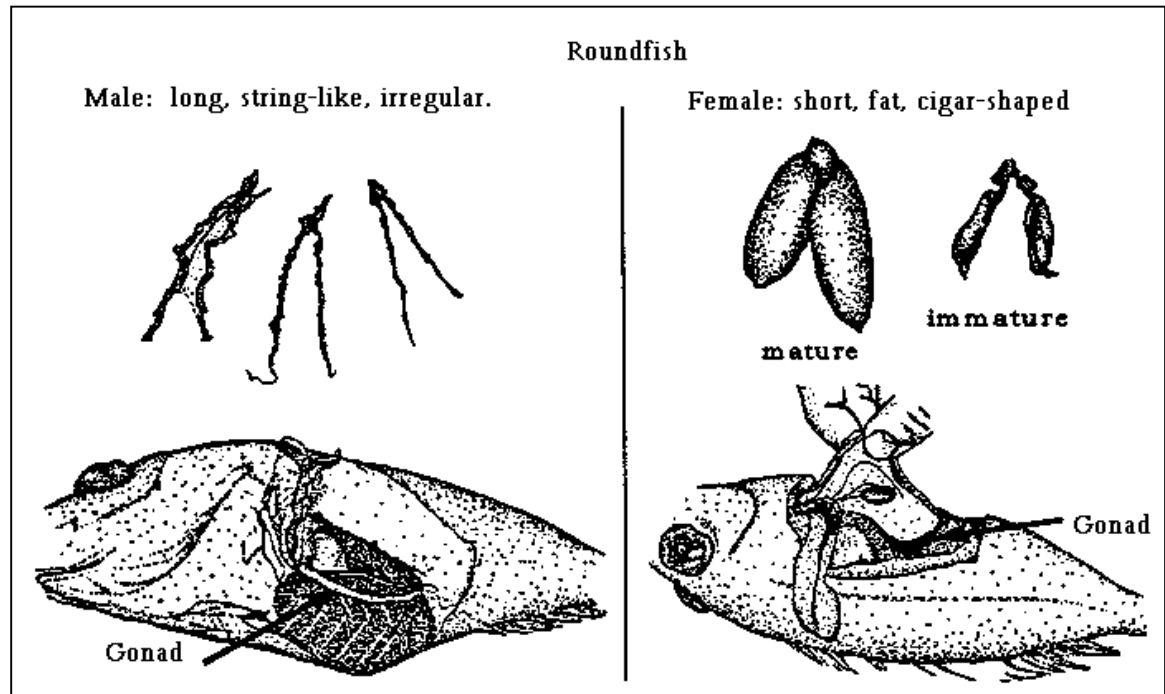


Figure 6- 5: Sexing Roundfish

Sexing Sablefish

The gonads of sablefish are very different from all other roundfish. They are located directly on the backbone, forward toward the head of the fish. Cut up from the anus to the collar to get a clear view. Alternatively, make a cut down from the lateral line to the belly, just behind the operculum, and then another insertion along the belly to the mid-point. Remove all the other organs from the visceral cavity and peer at the backbone area near the posterior of the cavity to look for the gonad tubes. Both females and males have fleshy smooth tubes which are cream or pink in color. Mature fish have liver colored gonad tubes while immature fish will have nearly see-through ribbons. There are no reliable differences in color or texture to differentiate between non-ripe males and females.

To differentiate between the sexes, probe the gonads lobes apart and count the lobes. Males will have four lobes and females will have two lobes. Be cautious when checking the number of gonad lobes. When mature, the ovaries may have a partial fold through each of the two lobes, giving a false impression of four lobes. Cut across the gonad strands to distinguish the true number of lobes.

Also be sure to look at the **posterior** part of the gonad. The lobes of males are fused at the anterior point of the gonad. Since the lobes will be fused anteriorly, they will always look like two lobes at this location and can be mistaken for female gonads.

Sexing Rockfish

Rockfish gonads are found near the backbone in the visceral cavity. Cut from the anus up to the collar and pull aside the intestinal organs. Be careful when doing so as the scales on rockfish are large and tend to make cutting difficult. Trace the gonad strings from the anus upwards until the paired organs are visible. There will always be two strings near the anus that have to be traced back before the sacs can be found. Sometimes there is another structure directly at the anus that appears to be a single gonad sac, but do not assess this as the sex organ! Always follow the string-like tubes up to the paired gonads.

The ovaries will be elongate ovals with granular insides. They will be pink, orange, yellow, or white. The two sacs will have smoothly rounded sides, as opposed to the male testes that have a three-sided, triangular shape in cross-section. If immature, look closely or cut the gonad open to see the granular insides that identify it as female. Rockfishes are live spawners, therefore, spawning females will have larvae in the cavity.

Rockfish testes are cream colored or pink, elongate (5 times as long as they are wide) and smooth in texture. They have three “edges” to the tubes. Instead of a rounded oval tube, testes look triangular in cross section due to the distinct edges. Testes will look like flat tubes when immature, but when examined closely you will see the sharp edges and the triangular shape.

Though you may notice external structures at the anus that seem sexually dimorphic, never sex rockfish using external characteristics. It is too easy to judge an immature male as a female or a huge female as a male when using external characteristics.

Sexing Flatfish

Flatfish gonads are paired, are located posterior to the visceral cavity and extend just under the flesh on both sides of the fish. If the flatfish has an anal spine, the gonads will begin just behind this spine. On the eyeless side of the fish, cut from the anal spine back toward the tail of the fish.

Lift the skin flap and check for a triangular shaped gonad.

Female flatfishes have elongate triangular ovaries that extend from behind the anal spine almost to the tail when mature. When immature, the ovaries are shaped like equilateral triangles with one corner shaped like a smoothly rounded tube extending slightly back toward the tail (the triangle looks like a funnel in shape). The color will be pink (spent, immature) or orange (ready to spawn). Ovaries always have rounded edges on the triangular gonad (See Figure 6-6).

Male flatfishes have a white, equilateral triangle shaped gonads. Unlike the female fish, the male's gonad is not elongated towards the posterior end. Immature males have a small crescent moon shaped, tan colored gonad lying directly at or slightly behind the anal spine. All male flatfishes have sharp edges on the triangular gonad (See Figure 6-6). Lift the gonad with a knife or scalpel and examine the sides of the triangular gonad to distinguish the sharp edges (male) or rounded sides (female). This technique works even on an immature flatfish.

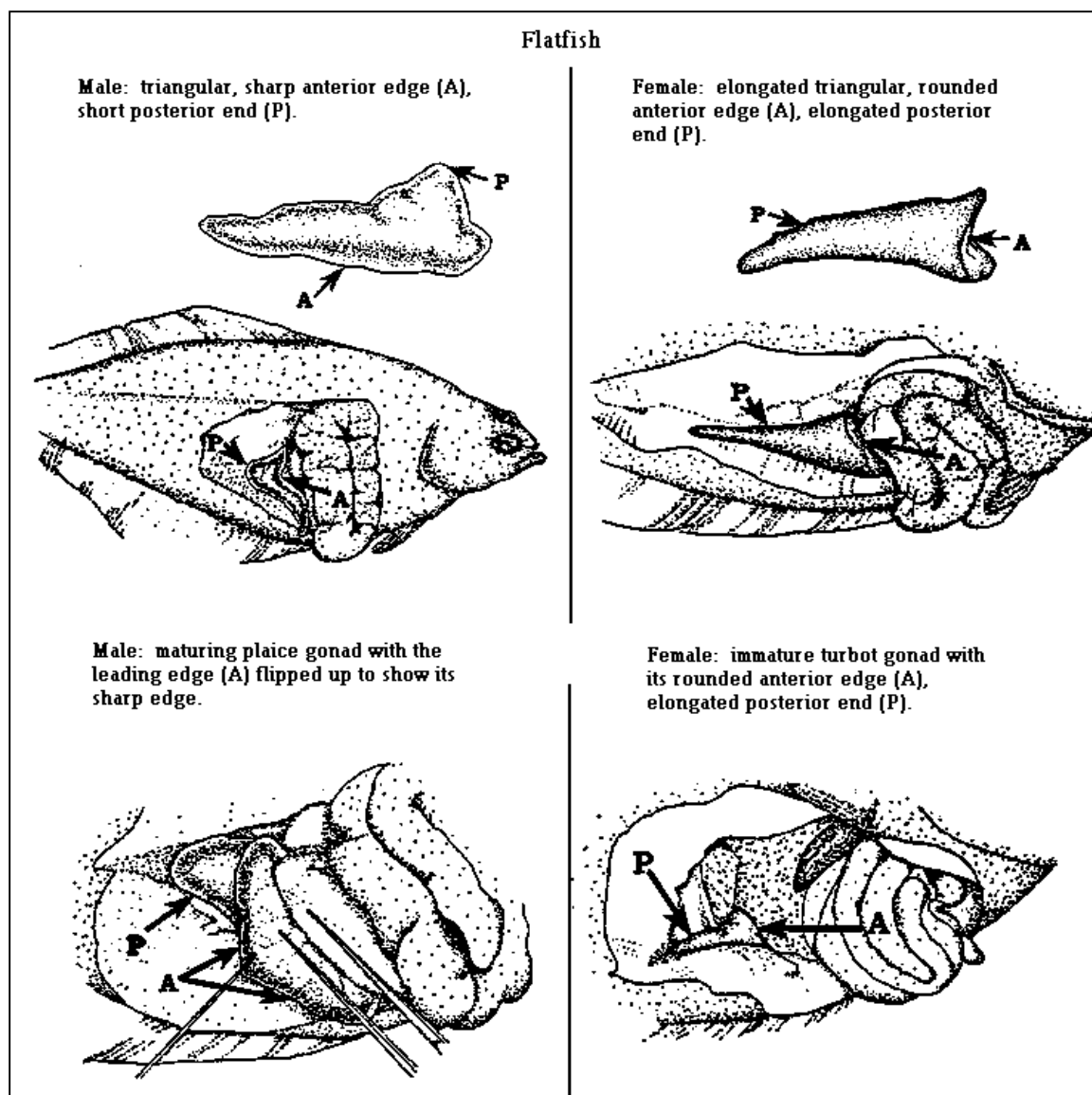


Figure 6- 6: Sexing Flatfish

Sexing Salmon

Salmon gonads are far forward in the body and immediately under the backbone. Cut into the salmon about mid way up from the belly (~1 cm below lateral line) and just behind the pectoral fin. Cut backward and down (about a 60 degree angle) until the top of the peritoneal cavity near the head is revealed. The gonads will be two long tubes lying parallel to the backbone. Females, mature and immature, will have tubes containing granular eggs in sacs that are pink, yellow, or orange. Mature males will have smooth textured tubes of white or cream color. Immature males will have translucent white tubes that appear empty.



Figure 6- 7: Otolith

Otoliths

Data Collection Guidelines

1. Collect otoliths from tagged sablefish.
2. Collect otoliths for high priority rockfish species.
3. Collect additional otoliths at the request of the WCGOP.

Otolith Location

The otoliths are located ventrally to either side of the brain tissue, just above where the pre-operculum is located. The common methods of cutting into a fish's head to remove the pair of otoliths are a vertical cut through the head above the pre-operculum or a horizontal cut through the head just above the eyes. The easiest method to use for most fish is to make a vertical cut down through the top of the head to the otolith pocket. This pocket is located at the two points on either side of the fish's head at which an imaginarily extended lateral line would meet the pre-operaculum bone.



Figure 6- 8: Taking Otolith

Species with tiny otoliths are best cut using the horizontal technique. Cutting to the correct point will break open the otolith cavities (one on each side of the brain) exposing the white, calcareous otoliths (See Figures 6-7 and 6-8).

Broken Otoliths

Otoliths are fairly fragile and must be in good condition to be read accurately.

Before collecting otoliths that will be used as part of a scientific collection, collect a variety of fish sizes and practice removing the otoliths. Try a variety of cuts and knife sizes to get comfortable with the angle and amount of pressure required. Field coordinators are available to suggest alternate techniques in cases where otoliths are consistently being broken.

Some otoliths may break or be cut accidentally during at sea collection. If both pieces are present, keep samples with otoliths that have a single break. Discard samples with a shattered otolith or with only one otolith.

Collecting Otoliths in General

Collect otoliths using the following procedure:

1. Firmly grasp the fish by putting thumb and forefinger into the eye sockets or grasp the fish just behind the head, holding it dorsal side up.
2. Position the knife on the top of the head and bear down on the knife with even pressure to cut through the headbone (See Figure 6-9). Pay attention to the amount of pressure being applied. As soon as the cutting gets easier, ease off pressure on the knife to avoid slicing through the otoliths.
3. Break the head open with two hands.

4. Remove the otoliths from the cavity with a pair of forceps.
5. Carefully clean the otoliths by rubbing them between your fingers in water, or on a wet sponge or cloth to remove slime and tissue.
6. Dry the otoliths as much as possible and place the pair of otoliths in a vial (only one pair of otoliths per vial). It is important to get the otoliths clean and as dry as possible before storing them to prevent their rotting.
7. Record the weight, sex, length, and species of the fish on the Biospecimen Form. Also record a Dissection Type of "1" for otoliths and record the bar code number from the otolith vial.

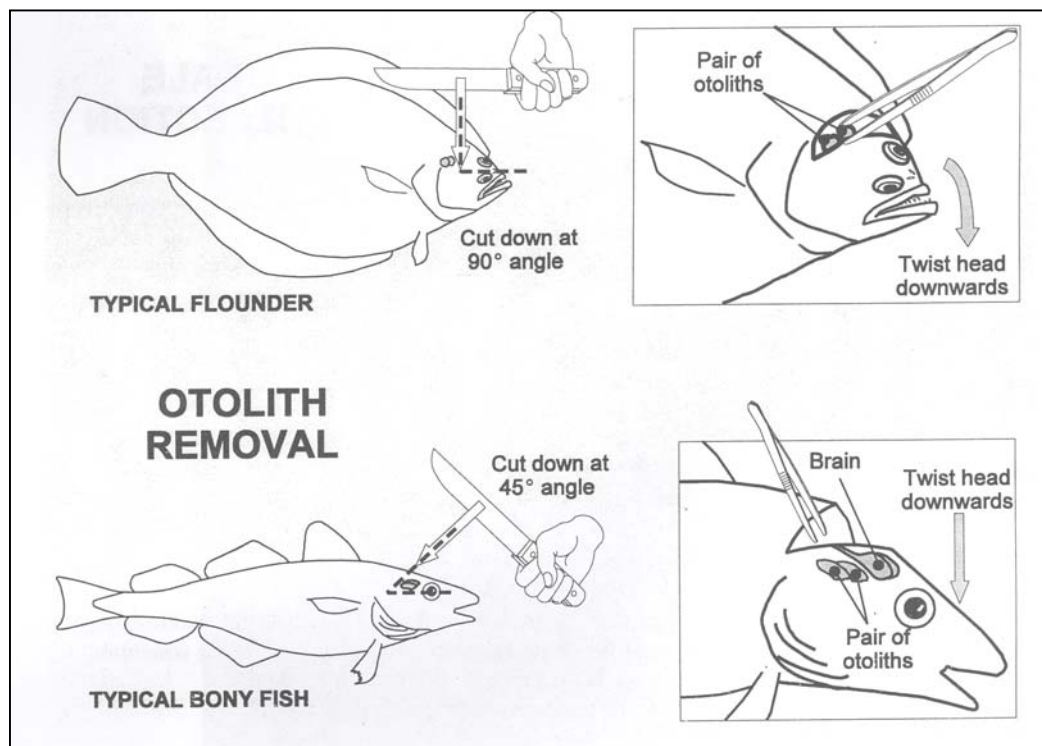


Figure 6- 9: Otolith Removal

Collecting Otoliths from Sablefish

Sablefish have very tiny otoliths. Employ a horizontal cut when working with this species (See Figure 6-10).

Collect sablefish otoliths using the following procedure:

1. Firmly grasp the fish's head.
2. Make a horizontal slice into the head just above the eye. Stop slicing when the knife is just before the preopercle.
3. Make a second vertical cut down into the head until the level of the first cut is reached.
4. Remove the wedge of cut skull. If the cut is correct, no blood should flood the cavity and the brain tissue should be visible.
5. Grasp the brain tissue with forceps and pull it out or peel it back from the cavity.
6. On either side of the brain cavity there is a fluid-filled pocket containing an otolith. Insert forceps into the pockets, to remove the bony structures floating within the fluid.
7. Carefully clean the otoliths by rubbing them between your fingers in water, or on a wet sponge or cloth to remove slime and tissue.
8. Dry the otoliths as much as possible and place the pair of otoliths a vial (only one pair of otoliths per vial). It is important to get the otoliths clean and as dry as possible before storing them to prevent their rotting.

9. Record the weight, sex, length, and species of the fish on the Biospecimen Form. Also record a Dissection Type of “1” for otoliths and record the bar code number from the otolith vial.

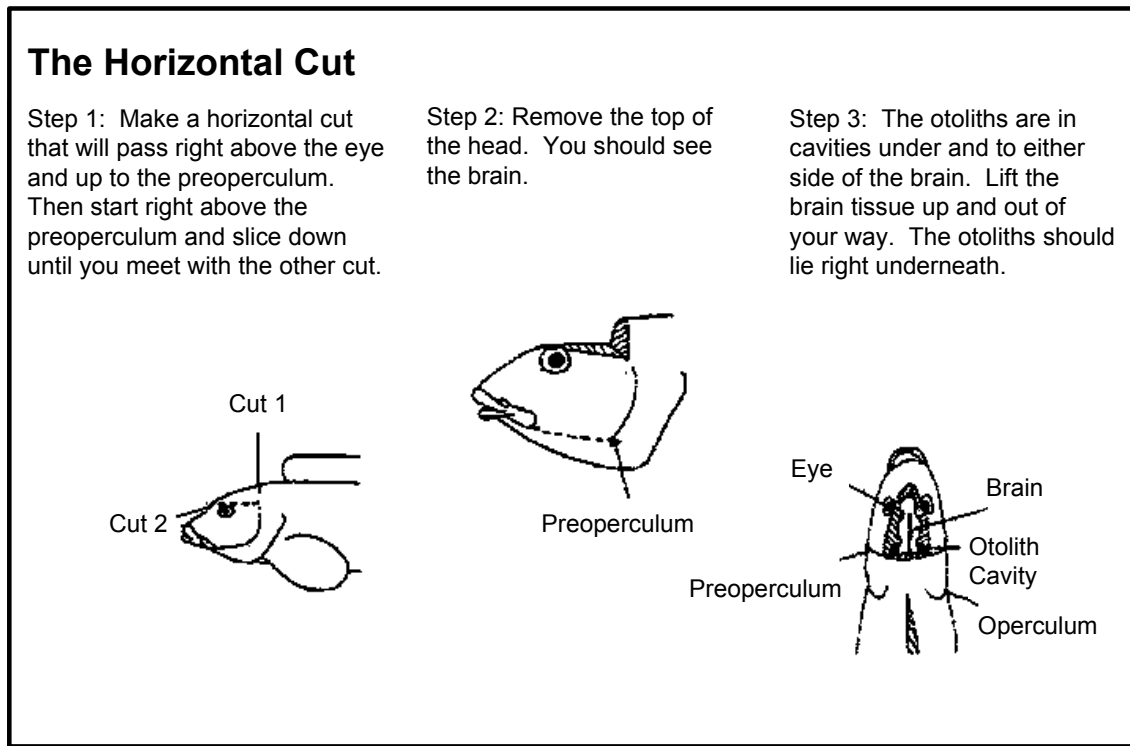


Figure 6- 10: Horizontal Cut Method

Pacific Halibut Viabilities

Data Collection Guidelines

1. Collect viabilities for DISCARDED Pacific halibut that have undergone NORMAL handling by the crew.
2. Always measure the length of Pacific halibut used for viabilities. Do NOT estimate the lengths.



Figure 6-11: Pacific halibut

Preparing to Collect Viabilities and Lengths from Pacific Halibut

In general, take viabilities and lengths for all discarded Pacific halibut in a haul/set. If you are unable to sample all of the Pacific halibut in a haul/set for viabilities and lengths, randomly select the individuals to use. A discussion of random sampling for biospecimens is contained earlier in this chapter.

The injury criteria and viability codes used to assess Pacific halibut viabilities vary by gear type. Make sure to use the correct set of criteria and codes when making injury assessments. A list of injury keys and codes follows:

1. Trawl Gear – “Viability Criteria and Injury Key for Trawl Caught Pacific Halibut” (Appendix K)
 - E – Excellent
 - P – Poor
 - D – Dead
2. Pot Gear - “Viability Criteria and Injury Key for Pot Caught Pacific Halibut” (Appendix L)
 - E – Excellent
 - P – Poor
 - D – Dead

3. Hook and Line Gear -“Viability Criteria and Injury Key for Hook and Line Caught Pacific Halibut” (Appendix M)
 - MI – Minor
 - MO – Moderate
 - S – Severe
 - D - Dead

Pacific halibut are often quite large and will be longer than the 100 cm length strips (See Figure 6-11). Prepare to take viabilities and lengths by offsetting the length strip by 20 or more centimeters. On the length strip, replace the M (male) and F (female) markings with the set of viability codes applicable to the gear type being used. As each Pacific halibut is measured and checked for injuries, a hatch mark will be recorded on the length strip for the appropriate length and viability.

Collecting Viabilities and Lengths from Pacific Halibut

Pacific halibut viabilities should NEVER be guessed. Always have the Pacific halibut in hand when taking viabilities.

In addition, viabilities should reflect the normal handling of Pacific halibut by the crew. If the vessel does not discard fish immediately, do not take the viabilities until the crew is preparing to discard the fish. The purpose of taking viabilities is to ascertain the condition of the fish when it returns to the sea.

Collect Pacific halibut viabilities and lengths using the following procedure:

1. Closely examine the Pacific halibut on both sides for injuries.
2. Use the appropriate Pacific halibut injury key to assign a viability code to the fish. Injury keys are located in the appendices as follows: Trawl (Appendix K), Pot (Appendix L) or Hook and Line (Appendix M).
3. Measure the fork length of the Pacific halibut by laying it directly on the plastic length strip. Never hold the tape measure over the top of the fish and “sight down” or take a curvilinear length as both of these methods introduce inaccuracies.
4. Place a hatch mark on the plastic length strip next to the appropriate length and viability.
5. Record the length and viability on the Biospecimen Form.



Salmon Scales

Data Collection Guidelines

1. Collect scales from the first five individuals of each salmon species encountered during your contract first contract, and the first two individuals encountered during each subsequent contract..
2. Collect scales for any salmon where you are unsure of the species.
3. Collect scales for all salmon where external tags or snouts are collected.
4. Collect additional scales at the request of the WCGOP.

Collecting Salmon Scales

Salmon lose scales easily and lost scales are replaced with regenerated scales. These regenerated scales and lateral line scales are unusable for aging purposes. Always collect at least 5 scales and never collect scales from the lateral line to be sure the scales are useable.

Collect salmon scales using the following procedure:

There are 8 species of salmonids encountered in the Eastern Pacific:

King (Chinook)
Silver (Coho)
Sockeye (Red)
Chum (Dog)
Pink (Humpback)
Atlantic salmon
Steelhead (Sea-run Rainbows)
Cutthroat trout

1. Wipe the area on the fish where you plan to collect scales. This ensures no other fish scales will be mixed in with the individual's scales. It also removes slime, which causes scales to decompose in the scale envelopes.



TIP* Remember, salmon rub against many other fish, even other salmon of different ages and species. To insure accurate data, make sure scales are clean.

2. Collect scales midbody from the area just above or below the lateral line. See Figure 6-12 for preferred scale collection zones. Never collect scales that lie directly on the lateral line.
3. Pluck salmon scales out of the flesh using forceps or a knife. Try to minimize mucus on the scales by plucking rather than scraping. If taking scales from multiple fish, be sure to clean the forceps between fish.
4. Open a paper salmon scale envelope and wipe the scales inside. Make sure you collect at least five scales. Seal the envelope.

5. Check for clipped adipose fin. If the adipose fin is clipped, the salmon is likely a tagged fish. Collect the snout of the salmon following the instructions in the Tagged Fish section of this chapter.
6. Weigh the salmon.
7. Determine the sex of the salmon.
8. Measure the length of the salmon.
9. Record the weight, sex , length , species, trip number and haul number on the scale envelope.
10. Record the weight, sex, length, and species of the fish on the Biospecimen Form. Also record a Dissection Type of “2” for scales and the bar code number from the scale envelope.

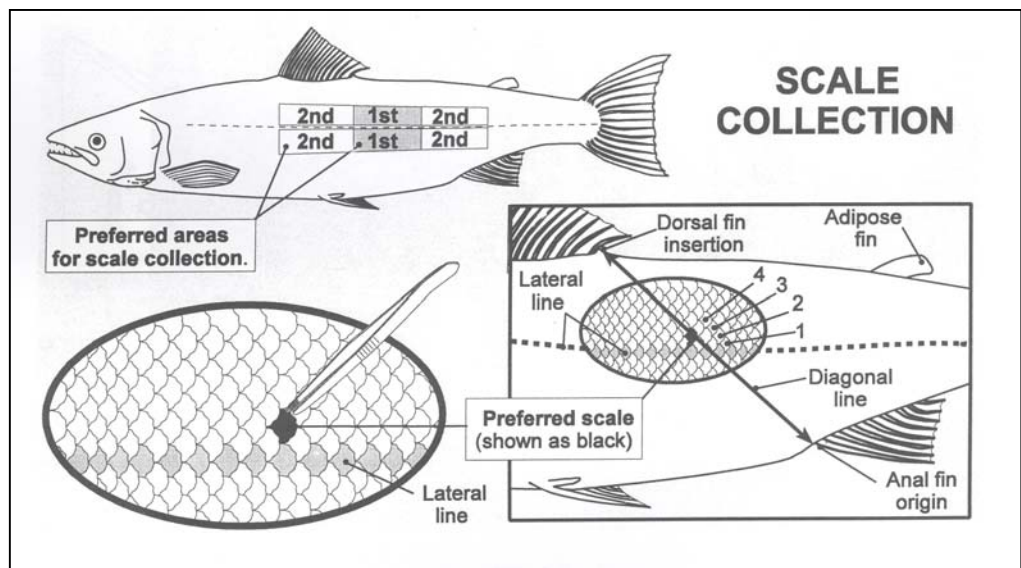


Figure 6- 12: Salmon Scale Collection

Tagged Fish

Data Collection Guidelines

1. Collect weight, length, sex, tag and tag data from all externally tagged fish.
2. Collect salmon weight, length, sex, scales, and snout from all salmon missing an adipose fin.

Preparing to Collect Tags

Inform the vessel crew that all tagged fish should be saved (See Figure 6-13). Species that have been tagged include Pacific cod, Pacific halibut, California halibut, pollock, sablefish, salmon, some rockfish species and shortspine thornyhead.

If vessel personnel provide an external tag or an externally tagged animal, write down the person's name and address to enable them to receive a reward for returning the tag. Observers cannot collect rewards for tags they submit. Observers who find tagged fish or crab should list the captain as the person who found the tagged animal.



Figure 6- 13: Tagged Sablefish

Collecting Tags

Tags for fish other than salmon are usually externally located on the dorsal surface or on the gill cover. Spaghetti tags are the most common type of external tag but some fish may have disc-shaped tags. If a tag is provided without the actual fish, collect as much information as possible from the crew member who gave you the tag including the tag collection date and location.

Tagged salmon usually have internal coded wire tags or PIT tags inserted into their snouts but some may have external disc shaped tags on their dorsal fins. Coded wire tags are about 1 mm in length and have a distinct code, usually a series of slashes at different intervals engraved on them. PIT tags are electronically coded tags that require a reader to decipher. Salmon tagged with coded-wire or PIT tags are identified by a missing adipose fin. Neither type of snout tag can be removed in the field, therefore, the entire snout must be collected and returned.

Collect biological information from tagged fish using the following procedure:

1. Remove the external tag or salmon snout. The collection procedure for salmon snouts follows this section.
2. Weigh the fish if possible.
3. Determine the sex of the fish.
 - In the case of LIVE Pacific halibut, do NOT attempt to sex the fish or remove the tag. Just record the tag number and other pertinent data.
4. Measure the length of the fish.

5. Collect otoliths from the fish.
 - In the case of LIVE Pacific halibut, do NOT attempt to collect otoliths.
 - In the case of sablefish with a blue spaghetti tag, collect the entire head and FREEZE it. The otoliths have been treated with a light sensitive chemical.
6. Collect scales if the fish is a salmon. The collection procedure for salmon scales is located earlier in the chapter.
7. Complete a Tagged Fish Form.
8. Record the weight, sex, length, and species of the fish on the Biospecimen Form. Also record a Dissection Type of “1” if otoliths have been taken, a “2” if salmon scales have been taken or a “3” if a snout has been collected. Record the bar code number from the otolith vial or scale envelope. Record external tag numbers in the Tag # column.

Collecting Salmon Snouts

Collect Salmon Snouts using the following procedure:



Figure 6- 14: Removed Salmon Snout

1. Make a cut one centimeter behind the eye, down through the head to the base of the upper jaw. The lower jaw does not need to be included since tags are placed in the upper snout. (See Figures 6-14 and 6-15)
2. Place the snout in one of the issued plastic bags and put several handfuls of table or rock salt in the bag. If no salt can be found, freeze the snout or place it in the hold with the retained fish.

3. Complete a Specimen Collection Label and include the set/retrieval location on the back of the label. Place the label inside the bag with the snout.
4. Periodically, drain off any liquid that accumulates in the bag and change the salt.

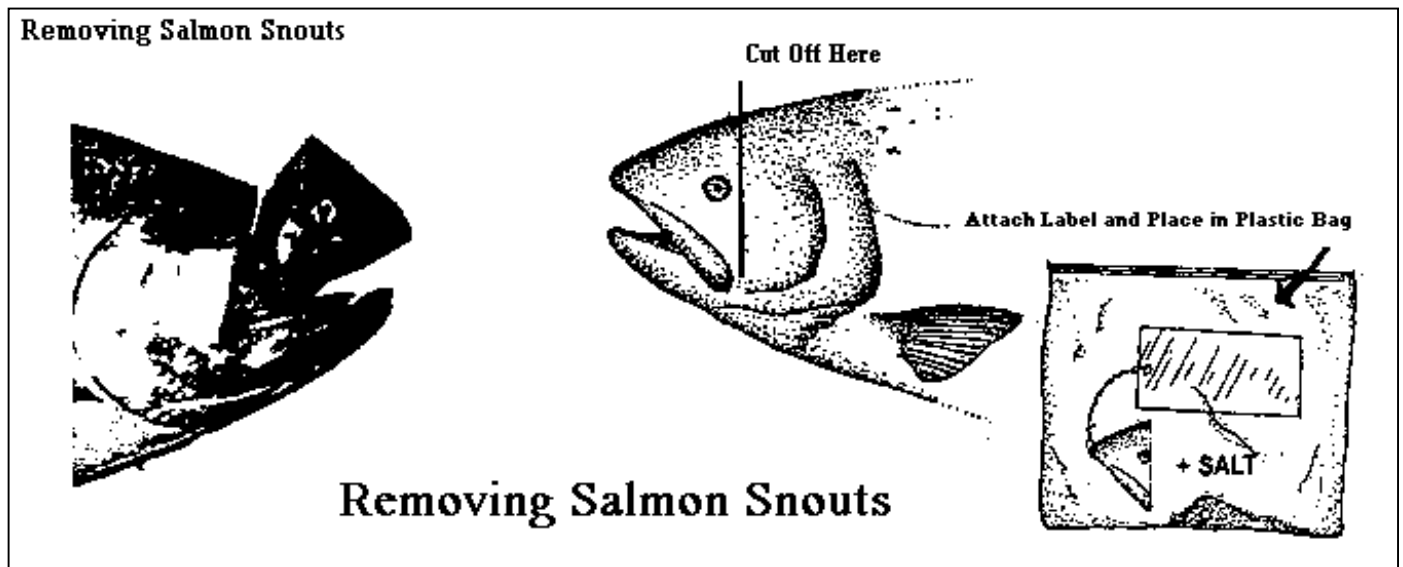


Figure 6- 15: Removing Salmon Snouts

Collecting Fish Specimens

Periodically observers will be asked to collect fish for training and other purposes. Observers are also required to bring in any fish that they are unable to identify at sea. When collecting fish adhere to the following procedures:

1. Place fish in a zip-lock bag. Do not include more than three fish per bag, and then only include fish of the same species.
2. Complete a Specimen Collection Label and place the label inside the zip-lock bag with the fish. If it is a specimen where the identification is uncertain, be sure to include the name used to enter the fish in the database.
3. Freeze the specimen as soon as possible. If the specimen cannot be frozen at sea, keep it on ice.
4. After returning to port, make arrangements to send the specimen to your coordinator, debriefer or to the Newport office. Always call ahead of time to insure that the specimens do not arrive when there is nobody present to receive them.

V. Data Collection Forms

There are four data collection forms to use when collecting biospecimen information.

1. Length Frequency Form
 - Use this form to record species, sex, and length data when only this information is being collected. Data can be recorded for individual fish or for groups of fish with identical species, sex and length values.
2. Biospecimen Form
 - Use this form to record data for individual fish when any information beyond species, sex, and length has been collected.
 - Use this form to record Pacific halibut viabilities.
 - Use this form to record data when otoliths, scales, snouts or tags have been collected.
3. Tagged Fish Form
 - Use this form to record data for tagged fish.
4. Specimen Collection Label
 - Use this form to record data when salmon or sablefish snouts have been collected.
 - Use this form to record data when whole fish/inverts have been collected.

Record data on either the Length Frequency Form or the Biospecimen Form. **NEVER** record data on both forms or the fish will be counted twice.

Length Frequency Form Instructions

Complete the Length Frequency Form for fish when only length and/or sex information is taken and no tags or dissections are collected. Fish should be grouped together whenever the species, length and sex for all the fish in the group are the same. An example of the form is included as Figure 6-16.

- **Haul Number** – Record the number of the haul that the sample came from.
- **Date** – Record the date as MM/DD/YY.
- **Trip Number** – Record the trip number generated by the database system.
- **Coast Guard Number** – Record the USCG vessel number on limited entry trawlers and fixed gear vessels (if they have one). Do not record anything in this field if you are on an open access vessel or a limited entry fixed gear vessel that does not have a USCG number.
- **Catch #** - Record the number that corresponds to the catch category on the Catch Form.
- **Catch Category** – Record in capital letters the catch category the species is in as recorded on the Catch Form.
- **R or D** – Record whether the sample came from an **R** – Retained or **D** – Discarded catch category.
- **Species Name** - Record the **common name** of the species the length frequencies were taken from. This column must be filled in with the species name. Do not only enter the species code! The common name listed on the paperwork must match the common name used in the database.

- **Species Code** - Record the species code of the corresponding species. See Appendices A and B for lists of species and species codes.
- **Method** – Record the Biological Sampling Method used to obtain fish for length frequencies.
 - 6 - Outside and Nonrandom
 - 7 - Outside and Random
 - 8 - Inside and Nonrandom
 - 9 - Inside and Random
- **Sex** – Record **M** – Male, **F** – Female, or **U** – Unknown (individuals where the sex cannot be determined). If you did not attempt to sex the individual, LEAVE THE COLUMN BLANK!
- **KP Length** – Sum up all of the length **by species** and note total of all lengths in the KP Length (keypunch length) column.
- **KP Frequency** - Sum up all of the frequencies **by species** and note total of all frequencies in KP Freq (keypunch frequency) column.
- **Length** – Record the length of the group of fish, in centimeters.
- **Freq** – Record the number of individual fish in each length group.

[illegible]

Figure 6-16: Length Frequency Form

Biospecimen Form Instructions

Complete the Biospecimen Form any time data beyond species, sex, and length are being collected on an individual fish. Complete this form when collecting Pacific halibut viabilities, otoliths, scales, snouts or tags. This form is also often used to record individual weights and lengths of fish caught in the Live Fish fishery. An example of the form is included as Figure 6-17.

- **Haul Number** – Record the number of the haul that the sample came from.
- **Date** – Record the date as MM/DD/YY.
- **Trip Number** – Record the trip number generated by the database system.
- **Coast Guard Number** – Record the USCG vessel number on limited entry trawlers and fixed gear vessels (if they have one). Do not record anything in this field if you are on an open access vessel or a limited entry fixed gear vessel that does not have a USCG number.
- **Catch #** - Record the number that corresponds to the catch category on the Catch Form.
- **Catch Category** – Record in capital letters the catch category the species is in as recorded on the Catch Form.
- **R or D** – Record whether the sample came from an **R** – Retained or **D** – Discarded catch category.
- **Species Name** - Record the **common name** of the species. This column must be filled in with the species name. Do not only enter the species code! The common name listed on the paperwork must match the common name used in the database.

- **Species Code** - Record the species code of the corresponding species. See Appendices A and B for lists of species and species codes.
- **Method** – Record the Biospecimen Sampling Method used to obtain fish for biospecimens.

6 - Outside and Nonrandom

7 - Outside and Random

8 - Inside and Nonrandom

9 - Inside and Random

- **Sex** – Record **M** – Male, **F** – Female, or **U** – Unknown (individuals where the sex cannot be determined). If you did not attempt to sex the individual, LEAVE COLUMN BLANK.
- **Viabilities** – Record the viability for **Pacific halibut ONLY**. Refer to Appendices K, L and M for viability criteria.

Trawl and Pot

D = Dead

P = Poor

E = Excellent

Hook and Line Gear

D = Dead

S = Severe

MO = Moderate

MI = Minor

- **Length** – Record the length of the individual fish in centimeters.
- **Weight** – Record the weight of the individual fish. Do not use extrapolated or halibut conversion weights.

- **Maturity Stage** – Record the maturity stage of the individual fish. This field should only be completed if special instructions from a coordinator are given.
- **Dissection Type** – Record the type of dissection that was taken. (Multiple dissection types can be recorded for an individual fish. Do NOT record all information for the fish multiple times – leave all fields except dissection type blank when recording extra dissections or it will appear that the dissections came from two different fish.)

1 – Otoliths
2 – Scales
3 – Snout
4 – Tissue

- **Barcode Number** – Record the barcode number of the vial, envelope, or other container that the dissected part was placed in.
- **Tag Number** – Record the tag number if the individual was tagged.
- **Comments** – Document any important information regarding the individual fish.
- **KP Length** – Sum up all of the lengths **by species** and note total of all lengths in the KP Length (keypunch length) column.
- **KP Frequency** - Sum up all of the frequencies **by species** and note total of all frequencies in KP Freq (keypunch frequency) column.

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Tagged Fish Form Instructions

Only complete the Tagged Fish Form for tagged fish. Attach the tag and otoliths directly to the form. An example of the form is included as Figure 6-18.

- **Trip No.** – Record the trip number generated by the database system.
- **Vessel ID No.** – Record the USCG number or state registration number (begins with CF, OR or WN) of the vessel.
- **Base Permit No.** – Record the Groundfish Permit number.
- **Observer Name** – Record your first and last name.
- **Vessel Name** – Record the name of the vessel on which the tag was collected.
- **Captain (or reward recipient's) Name** – Record the name of the person who found the tag or to whom any reward will be given. If the observer finds the tag, record the name of the vessel skipper or as otherwise instructed by the skipper.
- **Address** – Record the address of the reward recipient.
- **Species** – Record the common name of the species from which the tag was collected.
- **Tag Prefix and Serial No.** – Record this data if discernible from the tag.
- **Tagging Agency** – Circle which agency/lab tagged the specimen as recorded on the tag (if discernible).

- **Time and Date of Capture** – Record the retrieval time of the haul/set as MM/DD/YY.
- **Capture Location** – Record the retrieval position (latitude and longitude) of the haul/set.
- **Sex and Maturity of Gonads** – Record the sex of the fish. Do not record maturity stage.
- **Length** – Record the fork length of the fish in centimeters.
- **Weight** – Record the weight of the fish in pounds.
- **Capture Depth** – Record the retrieval depth of the haul/set in fathoms.
- **Vessel/Gear Type** – Record what gear type was utilized when the fish was captured (bottom trawl, midwater trawl, pot, longline, etc.)
- **General Appearance** – Note condition of the body including any wounds, scars or abnormalities.
- **Condition of Tagging Wound** – Note condition of the area around tag (open wound, scarred over, etc).
- **Other Comments** – Note anything else unusual or pertinent to the tagged fish.

TAGGED FISH FORM	
Trip No: _____	Vessel ID No: _____
Observer Name: _____	
Vessel Name: _____	
Base Permit No: _____	
Captain (or reward recipient's name): _____	
Address: _____	

Species: _____	
Tag Prefix (often a two letter code and Serial No): _____	
Tagging Agency (circle one): Seattle Auke Bay Nanaimo Shimizu IPHC Other _____	
Time and Date of Capture: _____	
Capture Location (Lat and Long): _____	
Sex and Maturity of Gonads (immature, mature, spawning): _____	
Length (fork length in cm): _____	
Weight (total wt. In lbs): _____	
Capture Depth (fathoms): _____	
Vessel/Gear Type: _____	
General Appearance (poor body condition, good body condition):	
Condition of Tagging Wound (healthy healed tissue, open wound):	
Other Comments:	
Attach Tag or vial here (with tape):	

Figure 6-18: Tagged Fish Form

Specimen Collection Label Instructions

Complete the Specimen Collection Label when salmon or sablefish snouts have been collected or when a whole fish or invert has been collected. An example of the form is included as Figure 6-19.



Tip* - Before going to sea, take 10 – 20 specimen collection labels and place a WCGOP bar code sticker on the back of the each label while the labels are clean and dry.

- **Vessel Name** – Record the name of the vessel on which the specimen was collected.
- **Haul Number** – Record the haul number from which the specimen was collected.
- **Trip Number** – Record the trip number generated by the database system.
- **Date** – Enter the date that the haul/set was retrieved as MM/DD/YY.
- **Species Identification** – Record the common name of the species.
- **Entered As** – Record the species name entered into the database, if this differs from the above (e.g. you entered it as rockfish, unidentified but believe it was a canary rockfish).
- **Depth (FM)** – Record the retrieval depth of the haul/set in fathoms.
- **Length (cm)** – Record the length of the fish, in centimeters.

- **Weight (LB)** – Record the weight of the fish, in pounds.
- **Sex** – Record the sex of the fish (if applicable).
- **Observer Name** – Record your first and last name.
- **Bar Code Sticker** – When collecting snouts, be sure to affix a WCGOP bar code sticker to the back of the specimen label in order to uniquely identify the specimen.

<p>SPECIMEN COLLECTION LABEL West Coast Groundfish Observer Program DOC/NOAA/NMFS/NWFSC/FRAMD 2725 Montlake Blvd. Seattle, WA 98112 (use pencil ONLY!)</p>	
VESSEL NAME _____ TRIP NUMBER _____ SPECIES IDENTIFICATION _____ ENTERED AS _____ DEPTH(FM) _____ WEIGHT(LB) _____ OBSERVER NAME _____	HAUL NUMBER _____ DATE _____ LENGTH(CM) _____ SEX (if applicable) _____

Figure 6-19: Specimen Collection Label